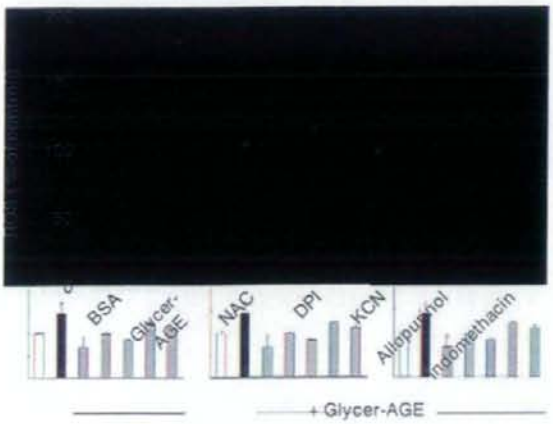

Fig. 2. Effects of advanced glycation end products (AGEs) on intracellular reactive oxygen species (*ROS*) generation Following exposure to glycer-AGE with or without *N*-acetylcysteine (*NAC*) for 24 h, ROS generation was quantified spectrophotometrically with 2',7'-dichlorofluorescein diacetate (DCFDA). To assess the source of ROS generation, the inhibitory effects of diphenylene iodonium chloride (*DPI*), potassium cyanide (*KCN*), allopurinol, and indomethacin were tested. Data are shown as means ± SEM from six independent experiments. $^{*}P < 0.01$ compared with nonglycated bovine serum albumin (*BSA*) as a control. $^{\#}P < 0.01$ compared with glycer-AGE alone

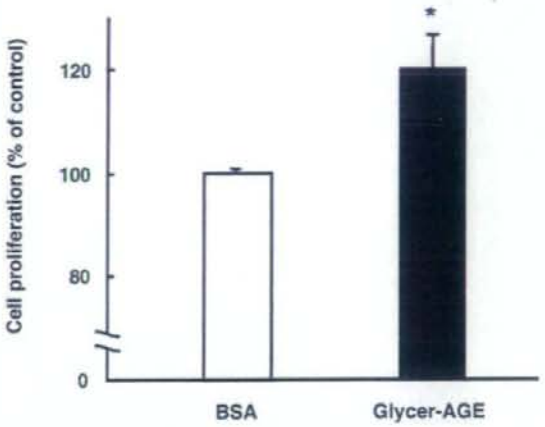

Fig. 3. LI90 cell proliferation following stimulation with AGEs. LI90 cells treated with glycer-AGE for 48 h were subjected to a cell proliferation assay with an MTS assay kit. Data are shown as means ± SEM from four independent experiments. $^{*}P < 0.05$ compared with nonglycated BSA as a control

chemicals: DPI, an NADPH oxidase inhibitor; KCN, an inhibitor of cytochrome oxidase in mitochondria; allopurinol, a xanthine oxidase inhibitor; and indomethacin, a cyclooxygenase inhibitor. These specific inhibitors did not cause morphological changes in LI90 cells, suggesting no cytotoxicities. Among these inhibitors, DPI and KCN significantly inhibited AGE-induced ROS generation, implying that the increased ROS were derived mainly from NADPH oxidase and the mitochondrial respiratory chain system.

Effect of AGEs on HSC proliferation

HSC proliferation is one of the key factors that provoke the progression of hepatic fibrogenesis. The MTS assay showed that stimulation by glycer-AGE significantly increased HSC proliferation, compared with the nonglycated BSA control (Fig. 3).

AGEs stimulate HSCs activation in a redox-sensitive manner

In addition to cell proliferation, activation of HSCs is a key process in hepatic fibrogenesis. Hence, we investigated the effects of AGEs on HSC activation. Transcripts encoding α-SMA and TGF-β1, both of which are established markers for HSC activation, were significantly upregulated by glycer-AGE (Fig. 4A and B). Moreover, mRNA of COL1A2, one of the major components of extracellular matrix, was also intensified by glycer-AGE (Fig. 4C). These changes were prevented by cotreatment with NAC.

Effect of AGEs on MCP-1 expression and secretion

MCP-1 is known to be secreted by HSCs and to amplify hepatic inflammation after liver injury.[19] To assess the possibility that AGEs influence hepatic inflammation through MCP-1 production, we investigated the expression of MCP-1 mRNA and secretion of MCP-1 protein. Glycer-AGE caused upregulation of MCP-1 mRNA (Fig. 5A) and stimulated secretion of MCP-1 protein into culture media (Fig. 5B); this secretion was markedly reduced by NAC.

Discussion

AGEs have been reported to accumulate in multiple tissues in diabetic patient and during normal aging, affecting tissue functions. We previously reported that six distinct AGE structures are elevated in the serum of diabetic patients,[20,21] and that among these, glycer-AGE exhibits neuronal toxicity in vitro.[22] We also have shown that glycer-AGE is prevalent in the serum of NASH patients.[5] In our preliminary experiments, we confirmed higher RAGE expression in liver biopsy tissue from NASH patients compared with tissues from healthy controls or NAFLD patients (data not shown). Hence,

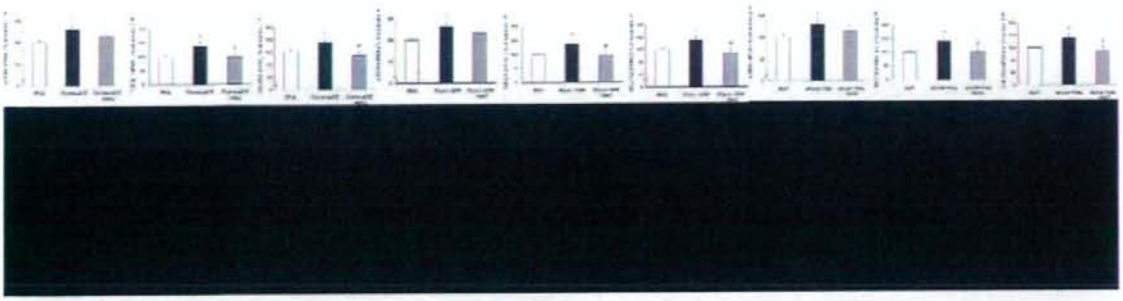

Fig. 4A–C. Effects of AGEs on the expression of mRNAs encoding hepatic fibrogenic markers. LI90 cells were incubated with glycer-AGE for 4 h, followed by quantitative real-time PCR measurement of mRNA encoding α-smooth muscle actin (*α-SMA*) (**A**), transforming growth factor β1 (*TGF-β1*) (**B**), and collagen type Iα2 (*COLIA2*) (**C**). Data are shown as means ± SEM from six independent experiments. $^{*}P < 0.01$; $^{**}P < 0.05$ compared with nonglycated BSA as a control; $^{\#}P < 0.01$ compared with glycer-AGE alone

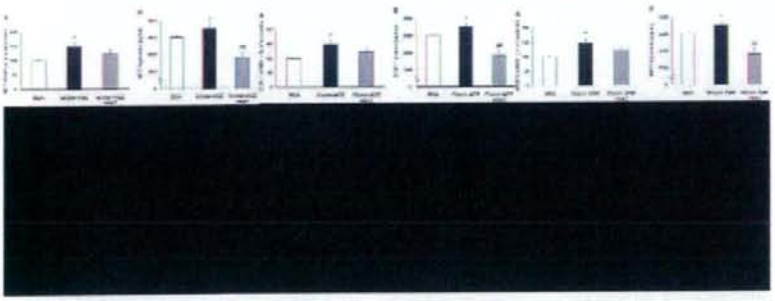

Fig. 5A, B. Monocyte chemoattractant protein 1 (*MCP-1*) production in LI90 cells following exposure to AGEs. **A** LI90 cells were stimulated for 4 h with glycer-AGE alone or in the presence of NAC. The level of MCP-1 mRNA was quantified by real-time PCR. **B** LI90 cells were stimulated for 48 h with glycer-AGE alone or in the presence of NAC. The level of MCP-1 protein in the conditioned media was quantified by enzyme-linked immunosorbent assay. Data are shown as means ± SEM from six independent experiments. $^{*}P < 0.01$; $^{**}P < 0.05$ compared with nonglycated BSA as a control; $^{\#\#}P < 0.05$ compared with glycer-AGE alone

we hypothesized that glycer-AGE might play an important role in the progression of NASH. In this connection, we examined the biological effect of glycer-AGE on human activated HSC line LI90.

Interestingly, a recent study reported that RAGE is present in rat primary HSCs and is upregulated during their transdifferentiation to myofibroblasts.[6] However, to date, no direct evidence of RAGE expression in human HSCs has been available. The present study demonstrated for the first time the expression of RAGE in human HSC line LI90 at both mRNA and protein levels.

We also provided evidence that treatment of HSCs with AGEs enhances intracellular ROS generation. This result is consistent with previous reports that the AGE–RAGE axis causes pathological conditions via induction of ROS generation in retinal pericytes, vascular endothelial cells, and mesangial cells.[1–3] Since ROS have been shown to stimulate HSC proliferation in vitro,[23] AGE-induced ROS generation might be one mechanism of liver damage in NASH patients. In fact, antioxidants such as betaine, *N*-acetylcysteine, and vitamin E have shown promising results on serum hepatic enzyme levels in patients with NASH.[24–26]

In this study, ROS generation in LI90 cell in response to AGEs was preferentially derived from NADPH oxidase and the mitochondrial respiratory chain system (Fig. 2). These findings are in agreement with the previous report that NADPH oxidase components are functionally expressed in HSCs and are the predominant contributor to angiotensin II-induced ROS generation in HSCs,[27] as well as to platelet-derived growth factor-induced proliferation of HSCs.[13] Furthermore, increased mitochondrial ROS formation in the liver has been

demonstrated in genetically diabetic *ob/ob* mice[28] and in rat NASH models.[29]

The finding in this study that AGEs intensified HSC activation in association with enhanced TGF-β1 adds new information to the mechanism of liver fibrogenesis under glucose intolerance. Recent studies have demonstrated that TGF-β is a potent inducer of both α1(I) and α2(I) collagen genes. In addition, a TGF-β-responsive element has been mapped to the promoter region of α2(I) collagen genes.[30] In keeping with this, the *COL1A2* gene was upregulated by AGE stimulation and accompanied by induction of TGF-β.

MCP-1 is a member of the CC subgroup of chemokines and is a potent chemoattractant of monocytes and T lymphocytes.[31] In addition to macrophages and endothelial cells, MCP-1 is secreted by activated HSCs, and it exhibits great involvement in the pathogenesis of NASH in animal models[10] and humans.[32] In this study, we demonstrated that stimulation of HSCs by AGEs led to upregulation of MCP-1, suggesting that AGE-induced MCP-1 secretion from activated HSCs elicits chronic liver inflammation during the progression of NASH.

Previous studies have indicated that activation of nuclear factor-κB (NF-κB) is induced by ROS in various nonphagocytic cells.[33] It has also been reported that AGEs activate NF-κB via an upstream signaling cascade, such as p38 mitogen-activated protein kinase in macrophages,[34] glomerular mesangial cells,[35] and endothelial cells.[36] Although further investigation is needed, we speculate from these observations that ROS generated by AGE–RAGE signaling may activate a transcription factor such as NF-κB, which results in activation of HSCs.

In this study, we utilized human HSC line LI90, which exhibits the characteristics of activated HSC when cultured in plastic dishes. Although we did not examine whether AGEs initiate the transdifferentiation from the quiescent to the activated phenotype, our findings that treatment with AGEs enhanced fibrotic markers as well as cell proliferation suggest that AGEs may play a role in the development of NASH, probably as a second hit.

In conclusion, we confirmed the expression of RAGE in human HSC line LI90. AGEs enhanced established fibrotic markers and proliferation of HSCs, which was associated with an increase of intracellular ROS generation via NADPH oxidase and the mitochondrial electron transport system. Together with the observation that AGEs are elevated in the serum of patients with NASH, these results suggest AGEs may be a second modulator in the progression of this disease. As antioxidants markedly retrieve the overexpression of fibrotic markers in HSCs, we speculate that the administration of antioxidants may represent a novel treatment strategy for NASH patients.

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HEPATOLOGY

Prospective study of short-term peginterferon- -2a monotherapy in patients who had a virological response at 2 weeks after initiation of interferon therapy

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Key words
adverse effects, clinical trial, efficacy, hepatitis C virus, peginterferon- -2a.

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Abstract

Background and Aims: Long-term interferon (IFN) therapy is effective in eliminating hepatitis C virus (HCV). However, it carries the risk of adverse effects and reduced quality of life. To assess whether short-term IFN therapy effectively eliminates HCV, we performed a prospective pilot study of pegylated (peg)IFN- -2a therapy for 8 or 24 weeks.

Methods: After excluding patients with high titers of genotype-1, 55 HCV patients received pegIFN- -2a. Patients who became negative for HCV-RNA at week 2 were allocated to either an 8-week (n = 19) or 24-week (n = 15) course of IFN. We evaluated the efficacy of and tolerance to IFN therapy.

Results: The sustained virological response rate was excellent in the two groups (8 weeks, 89.5% [17/19]; 24 weeks, 100% [15/15], respectively,). IFN dose reduction was required in one patient of the 8-week group, but in six patients of the 24-week group (P = 0.028). Treatment was completed by all patients of the 8-week group, but discontinued in five patients of the 24-week group (P = 0.011).

Conclusions: The 8-week IFN therapy is more tolerable than the 24-week therapy and had similar outcomes. Excluding the patients with high titers of genotype-1, we recommend switching to an 8-week course of pegIFN- monotherapy once patients show an ultra rapid virological response at week 2 from the start of IFN therapy.

Introduction

Hepatitis C virus (HCV).infection is a major cause of chronic liver disease with an estimated 170 million chronic carriers worldwide.[1] Chronic HCV infection is usually associated with liver cirrhosis (LC) and hepatocellular carcinoma (HCC).[2–6] In Japan, 60–70% of patients with HCC or LC are HCV carriers.[7] Antiviral therapy of interferon (IFN) is widely used for the treatment of chronic HCV infection and is assumed to prevent progression to LC and HCC, especially in patients who show a sustained virological response (SVR).

The reported total HCV-RNA elimination rate is approximately 30–40% in patients treated with conventional IFN monotherapy.[8–10] However, better results have been reported when pegylated (peg)IFN- is used in both naive patients and in those who fail to respond to or relapse after conventional IFN- monotherapy. In Japan, two kinds of pegIFN are available: pegIFN- -2a and pegIFN- -2b. PegIFN- -2b can be used with ribavirin, a purine nucleoside analog, in naive patients with genotypes 1 and 2 with a high viral load (>100 KIU/mL of HCV-RNA) or patients with any viral load in whom previous IFN treatment did not eliminate HCV-RNA. PegIFN- -2a has been used in Japan without ribavirin only since December 2003 because of health insurance restrictions. However, ribavirin combination therapy has been covered by public health insurance since March 2007 in Japan. The HCV elimination rate with pegIFN- -2b plus ribavirin combination therapy is up to 54% in patients with genotype 1.[11] Several investigators have reported that pegIFN and ribavirin combination therapy for a period of 24 or 48 weeks ensures a viral clearance in most patients with HCV genotypes 2 or 3 infection.[12,13] However, ribavirin combination therapy frequently causes anemia and should be carefully used in the elderly, anemic, or pregnant young patients, and in those who require long-term treatment.[14] Apart from patients with a high viral load of genotype 1, IFN monotherapy is also effective in HCV elimination even when used without ribavirin. Previous studies suggest that the SVR achieved with pegIFN- -2a is similar to that observed with pegIFN- -2a combined with ribavirin in patients with hepatitis C.[15,16]

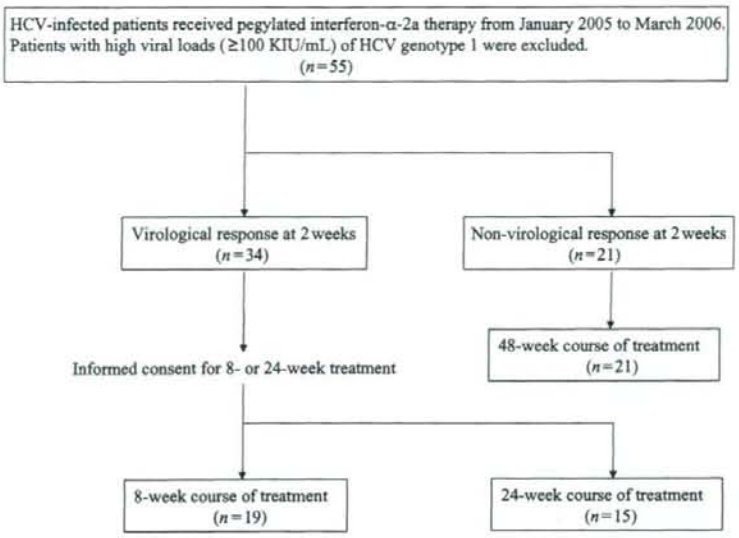

Figure 1 Flow diagram of the clinical trial. HCV, hepatitis C virus.

Although the tolerability of pegIFN is similar to that of the conventional IFN,[15] the 180 g dose of pegIFN- -2a therapy for 48 weeks is sometimes not tolerated by some patients. With the exception of those with high viral loads of genotype 1, the above regimen is expected to produce a high viral clearance rate, especially in patients with an early virological response. Several studies report the effectiveness of short-course IFN therapy (<24 weeks) for patients with an early virological response.[17–20] Therefore, a treatment duration of 48 weeks may be too long or more than sufficient for some patients, especially when one considers the undesirable adverse effects or the cost of treatment.

In the present study, we conducted a prospective controlled trial to compare the efficacy of an 8-week versus a 24-week course of pegIFN- -2a (180 g/time/week) for patients negative for HCV-RNA at 2 weeks after the initiation of therapy.

Methods

Patients

Between January 2005 and March 2006, a total of 55 HCV-infected patients received pegIFN- -2a therapy at Hiroshima University Hospital (Hiroshima, Japan) and its associated hospitals in Japan. Patients with high viral loads (≥100 KIU/mL) of HCV genotype 1 were excluded from this study because of their low SVR rate. Among the 55 patients, 34 consecutive patients who showed a rapid virological response at 2 weeks were enrolled in this study (Fig. 1). Eligible patients had antibodies to HCV, were positive for HCV-RNA at study entry, and had not received previous IFN therapy. They included 21 men and 13 women, with a mean age of 53 years (range, 21–71 years). Their HCV genotypes were 1b, 2a, and 2b with variant HCV-RNA (5.1–400 KIU/mL by a reverse transcriptase-polymerase chain reaction [RT-PCR]). All patients underwent liver biopsies within 12 weeks before the start of IFN therapy and were confirmed to have chronic hepatitis by histopathological examination. Patients with any other cause of liver disease including coinfection with hepatitis-B virus or HIV, alcoholic hepatitis, fatty liver, autoimmune hepatitis, or previous organ transplantation were excluded from this study.

Study design

This multicenter prospective controlled study compared the efficacy and safety of 8 weeks versus 24 weeks of pegIFN- -2a monotherapy in previously untreated patients with chronic hepatitis C who had a virological response at 2 weeks after the start of IFN. Patients with a virological response at 2 weeks were invited to sign a consent form accepting treatment with IFN for 8 weeks only. Those patients who refused consent received a 24-week course of treatment. The primary measure of efficacy was SVR, which was defined as undetectable HCV-RNA in the serum at 24 weeks after the cessation of treatment. All patients agreed to participate in the research protocol, which was approved by the hospital research ethics board, and gave written informed consent. The eligible patients received pegIFN- -2a (Pegasys, F. Hoffmann-LaRoche, Basel, Switzerland) at 180 g once per week subcutaneously, either for 8 weeks or 24 weeks, without ribavirin. Other patients who showed no rapid virological response at 2 weeks after the start of pegIFN- -2a were treated for 24–48 weeks.

All patients were evaluated in an outpatient setting for safety, tolerance, and efficacy every week during the IFN treatment. Blood count was checked just before the IFN injection every week. The qualitative detection of HCV-RNA was performed by a standardized qualitative RT–PCR assay (Amplicor HCV monitor v2.0; Roche diagnostics Co., Tokyo, Japan) at the first 2 weeks and every 4 weeks during and after IFN treatment. The primary efficacy end point for this study was defined as a disappearance of detectable serum HCV-RNA at week 24 after the completion of the IFN treatment.

Table 1 Patients' characteristics

	8-week group (n = 19)	24-week group (n = 15)
Age (years)	51† (21–71)	47† (25–58)
Sex (male/female)	14/5	7/8
Height (cm)	169† (147–178)	161† (139–178)
Weight (kg)	64.6† (40.6–85)	59† (47–92.4)
Body mass index (kg/m²)	23.4† (18.0–27.8)	21.5† (18.6–30.5)
Platelet count (10⁴/ L)	19.5† (9.6–30.7)	18.1† (8.9–31.7)
Alanine aminotransferase (IU/L)	55† (22–152)	60† (21–184)
-Glutamyl transpeptidase (IU/L)	25† (9–155)	47† (14–137)
Creatinine (mg/dL)	0.76† (0.6–0.9)	0.68† (0.38–0.85)
Total cholesterol (mg/dL)	160† (116–219)	154† (125–201)
Fasting blood glucose (mg/dL)	90† (72–104)	96† (84–115)
Diabetes mellitus	0	1
Hyaluronic acid (ng/mL)	24† (13–72)	78† (16–191)
HCV genotype (1b/2a/2b)	2/15/2	2/10/3
HCV-RNA (KIU/mL)	45† (5.1–370)	43† (5.3–400)
Fibrosis (F1/F2/F3/F4)	6/8/5/0	5/6/4/0

†Median. HCV, hepatitis C virus.

Statistical analysis

We compared the response to an 8-week course of pegIFN- -2a with that to a 24-week course of pegIFN- -2a. The ²-test and Fisher's exact test were used for comparisons of categorical variables between groups, while Student's t-test and the Wilcoxon test were used for continuous and ordinal variables as appropriate. P-values less than 0.05 were considered to indicate statistical significance. The JMP version 5.1 statistical software package (SAS Institute, Cary, NC, USA) was used for the statistical analysis of data.

Results

Baseline characteristics

Thirty-four patients who became HCV-RNA-negative at week 2 subsequently received either an 8-week course (n = 19) or 24-week (n = 15) course of 180 g pegIFN- -2a. The baseline characteristics of the two groups at the start of the IFN therapy are summarized in Table 1. None of the patients had LC, based on clinical, laboratory, and histopathological findings. Table 2 also shows the data of 21 patients with a non-rapid virological response at 2 weeks after the start of pegIFN- -2a. The pretreatment viral loads of non-rapid virological responders were significantly higher than those of rapid virological responders (P < 0.0001).

Tolerance of IFN therapy and adverse events

Among the 19 patients of the 8-week group, the dose was reduced by 50% (to 90 g of pegIFN- -2a) in one patient with SVR at 3 weeks due to a fall in platelet count. However, all other patients were able to complete the full 8-week course without discontinuation. In 15 patients of the 24-week course, the dose was reduced to half (90 g of pegIFN- -2a) in six patients due to neutropenia (n = 2; one patient at 8 weeks and one patient at 10 weeks), thrombocytopenia (n = 3; two patients at 9 weeks and one patient at 10 weeks) and epigastralgia (n = 1; at 14 weeks). Furthermore, IFN therapy was withdrawn in another five patients, including two patients at 8 weeks due to thrombocytopenia, two patients at 12 weeks due to generalized fatigue, and one patient at 18 weeks due to various neurological symptoms, such as hand numbness. Thus the proportion of patients who required a dose reduction was lower in the 8-week group than in the 24-week group (P = 0.028). Furthermore, the proportion of patients who completed the treatment was significantly higher in the 8-week group than the 24-week group (P = 0.011). We concluded that our patients with HCV could tolerate 8 weeks of IFN therapy better than 24 weeks.

Table 2 Characteristics of 21 patients who did not show a rapid virological response

Age (years)	51† (22–76)
Sex (male/female)	11/8
Height (cm)	164.5† (148–175.5)
Weight (kg)	58.5† (42.5–75)
Body mass index (kg/m2)	22.5† (16.9–27.3)
Platelet count (104/_L)	20.5† (12–28.6)
Alanine aminotransferase (IU/L)	93† (17–157)
-Glutamyl transpeptidase (IU/L)	39† (10–145)
Creatinine (mg/dL)	0.58† (0.5–0.96)
Total cholesterol (mg/dL)	158† (111–214)
Fasting blood glucose (mg/dL)	87† (68–119)
Diabetes mellitus	0
Hyaluronic acid (ng/mL)	45.6† (10–100)
HCV genotype (1b/2a/2b)	0/15/6
HCV-RNA (KIU/mL)	660† (40–830)
Fibrosis (F1/F2/F3/F4)	12/6/3/0

†Median. HCV, hepatitis C virus.

Biochemical and virological responses to therapy

With regard to the alanine aminotransferase (ALT) response to IFN therapy, all patients of both groups showed biochemical normalization at the end of treatment and at 6 months after the end of treatment. There was no difference in the sustained ALT response between the 8-week group and 24-week group. With regard to the virological response to IFN therapy, all patients of both groups exhibited a rapid decrease in HCV-RNA, reaching undetectable levels (HCV-RNA ≤ 100 copies/mL) by week 2. All patients had negative HCV-RNA levels at the end of treatment and none showed a null response. There was no significant difference in the rate of fall of the virological load between patients who had a sustained response and those who had a relapse, as discussed later. The proportions of patients who showed a SVR in the 8-week group and 24-week group were not significantly different (89.5% [17/19] and 100% [15/15], respectively [P = 0.195]). Two patients of the 8-week group had viral relapse after the end of treatment; one who had HCV genotype 2a with 50 KIU/mL pretreatment viral load relapsed at 12 weeks after the end of the treatment while the other had genotype 2b with 230 KIU/mL pretreatment viral load and relapsed at 8 weeks after the end of the treatment. The non-

rapid virological responders had a lower SVR rate. Eight (38%) patients showed SVR, 11 (52%) patients developed relapse after discontinuation of IFN, and two (10%) patients had no virological response.

Discussion

In Japan, pegIFN- -2a monotherapy has been covered by public health insurance since December 2003. The standard duration of treatment with pegIFN- -2a is 24 weeks for patients with low viral loads of genotype HCV-1 and any viral loads of genotype HCV-2 infection. Recent studies have reported that a treatment duration of more than 24 weeks in such cases does not increase the SVR rate.[11,13,21,22] Moreover, patients with early virological response seem to have a high rate of SVR.[23–25] In those patients, to reduce unnecessary exposure to treatment and its potential side-effects and to reduce costs, short-term IFN therapy has been used by several groups.[17–21] However, details of the IFN regimen differ from those of others and there are no studies that use short-term of pegIFN- -2a treatment. We therefore conducted a prospective pilot study on the efficacies of an 8-week and 24-week pegIFN- -2a regimen for patients with low viral titers of genotype HCV-1 and any viral titers of genotype HCV-2 who exhibited a virological response at 2 weeks after the initiation of IFN. In our study, patients with a relatively low viral load before the start of the IFN therapy tended to have a very early virological response.

Our results demonstrated that the virological response to the 8-week treatment (89.5% [17/19]) was excellent and was similar to the 24-week course (100% [15/15]). This high SVR rate of 8-week pegIFN- -2a monotherapy seems as high as that reported in another short course study of 14-week pegIFN plus ribavirin combination treatment for patients with HCV genotype HCV-2 or HCV-3.[26] This high SVR rate of the 8-week course of pegIFN- -2a may be associated with a rapid viral disappearance. Several studies have indicated that negative HCV-RNA at week 2 after the commencement of IFN is a predictor of SVR.[19,20,27,28] Therefore, for patients with a low HCV-1 viral load or those with HCV-2 infection with any viral load, we recommend switching to an 8-week course of pegIFN- -2a monotherapy once they show an ultra rapid virological response, that is, negative HCV-RNA at week 2 from the start of IFN therapy. Furthermore, a longer course of IFN therapy with or without ribavirin can be prescribed when HCV-RNA becomes positive after discontinuation of the 8-week course of IFN therapy.

Although in our study all of the patients in the 24-week course showed SVR, it seems that 24 weeks is a long treatment period for those patients who become negative for HCV-RNA by week 2 of treatment to ascertain SVR. Our results showed that all patients in the 8-week course completed the course to the end of treatment. However, 33% of the patients of the 24-week course did not continue their treatment to the end of the course. Because patients tend to adhere to shorter regimens, which are also better tolerated than longer treatment regimens, a shorter exposure will probably translate into a better benefit–risk ratio in patients with early virological response.

Our study identified two relapsers among patients of the 8-week course after discontinuation of pegIFN- -2a therapy. These two patients had a negative history of exposure to new HCV infection. One patient who had genotype HCV-2b and a high pretreatment viral load (HCV-RNA: 230KIU/mL) relapsed at 8 weeks after the discontinuation of IFN therapy, while the other who had genotype HCV-2a and a low pretreatment viral load (HCV-RNA: 50KIU/mL) relapsed at 12 weeks after the discontinuation of IFN therapy. These two patients could have SVR after additional IFN therapy for 24 weeks (one patient; pegIFN- -2a monotherapy, one patient; pegIFN- -2b and ribavirin combination therapy). We could not identify a definite factor associated with SVR or relapse. Although pretreatment factors, like genotype, viral load, and grade of fibrosis can be used to predict the mean treatment outcome for study cohorts, they are often of limited value in individual patients.[29,30]

As mentioned earlier, a short course of pegIFN- -2a therapy for 8 weeks could be recommended in those patients who show an ultra rapid virological response at week 2 after the initiation of IFN therapy. Although the study by Shiffman et al.[31] demonstrates the inferiority of a shorter regimen in a large-scale, randomized, controlled study, the characteristics of their patients were largely different from those of our study, including racial difference (mostly Caucasian patients versus Japanese, a heavier body weight for the Caucasians versus Japanese patients) and differences in pretreatment viral load (variable and higher HCV-RNA level versus relatively low viral load in our patients). Patients' selection was also different between the two studies; our study was carried out only in cases negative for HCV-RNA at 2 weeks after the start of IFN compared to their randomized study, irrespective of a rapid virological response.

In conclusion, patients chronically infected with low titers of HCV-1 and those with HCV-2, regardless of their viral loads, who achieve an ultra rapid virological response, that is, HCV-RNA negativity at week 2, can receive only 8 weeks of pegIFN- -2a monotherapy without compromising the chance of SVR. The results of our prospective study are encouraging, although the study population was small and was based on non-randomized methodology. The data of the present study are not conclusive for patients with very high pretreatment viremia who might achieve a rapid virological response or for those patients who do not achieve a rapid virological response. Further clinical trials are required to optimize the treatment duration in these patients.

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Susceptibility of Chimeric Mice with Livers Repopulated by Serially Subcultured Human Hepatocytes to Hepatitis B Virus

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We previously identified a small population of replicative hepatocytes in long-term cultures of human adult parenchymal hepatocytes (PHs) at a frequency of 0.01%-0.09%. These hepatocytes were able to grow continuously through serial subcultures as colony-forming parenchymal hepatocytes (CFPHs). In the present study, we generated gene expression profiles for cultured CFPHs and found that they expressed cytokeratin 19, CD90 (Thy-1), and CD44, but not mature hepatocyte markers such as tryptophan-2,3-dioxygenase (TO) and glucose-6-phosphatase (G6P), confirming that these cells are hepatic progenitor-like cells. The cultured CFPHs were resistant to infection with human hepatitis B virus (HBV). To examine the growth and differentiation capacity of the cells *in vivo*, serially subcultured CFPHs were transplanted into the progeny of a cross between albumin promoter/enhancer-driven urokinase plasminogen activator-transgenic mice and severe combined immunodeficient (SCID) mice. The cells were engrafted into the liver and were able to grow for at least 10 weeks, ultimately reaching a maximum occupancy rate of 27%. The CFPHs in the host liver expressed differentiation markers such as TO, G6P, and cytochrome P450 subtypes and could be infected with HBV. CFPH-chimeric mice with a relatively high replacement rate exhibited viremia and had high serum levels of hepatitis B surface antigen. *Conclusion:* Serially subcultured human hepatic progenitor-like cells from postnatal livers successfully repopulated injured livers and exhibited several phenotypes of mature hepatocytes, including susceptibility to HBV. *In vitro*–expanded CFPHs can be used to characterize the differentiation state of human hepatic progenitor-like cells. (HEPATOLOGY 2008;47:435-446.)

Abbreviations: 9MM, 9-month-old Caucasian male; 10YF, 10-year-old Caucasian female; 12YM, 12-year-old Asian male; 16YF, 16-year-old Asian female; AAT, α1-antitrypsin; AFP, α-fetoprotein; ALB, albumin; BGP, biliary glycoprotein; BrdU, 5-bromo-2′-deoxyuridine; CFPH, colony-forming parenchymal hepatocyte; CK, cytokeratin; G6P, glucose-6-phosphatase; h, human; HBsAg, hepatitis B surface antigen; HBV, hepatitis B virus; CYP, cytochrome P450; m, mouse; MDR, multidrug resistance protein; MRP, multidrug resistance-associated protein; PH, parenchymal hepatocyte; RI, replacement index; RT-PCR, reverse-transcription polymerase chain reaction; SH, small hepatocyte; TO, tryptophan-2,3-dioxygenase; uPA, urokinase plasminogen activator.

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Studies using rodents with damaged livers have shown that parenchymal hepatocytes (PHs) have great growth potential. When mouse (*m*) hepatocytes were transplanted into the livers of albumin promoter/enhancer-driven urokinase plasminogen activator (uPA)-transgenic mice,[1] they engrafted and repopulated the host liver. Serial transplantation experiments using *m*-hepatocytes in mice with tyrosinemia showed their enormous growth capacity.[2] The replicative potential of rat hepatocytes has also been demonstrated by transplanting them into the partially hepatectomized liver of a retrorsine-treated rat,[3] and uPA-transgenic mice crossed with severely immunodeficient mice, such as severe combined immunodeficient (SCID)/beige mice,[4] SCID mice,[5,6] or recombination activation gene 2 knockout mice[7] have been used to show the growth potential of human (*h*)-hepatocytes. When transplanted into uPA/SCID mice, PHs from a human juvenile male grew in the host liver to a level at which the proportion (replacement index) of the area of repopulated *h*-hepatocytes to the total number (host and donor) of hepatocytes reached 96% at 64 days posttransplantation.[5] Such *h*-hepatocyte–chimeric mice have been used to study the pharmacological responses of *h*-hepatocytes[5] and to investigate *h*-hepatitis viral infections.[4,6-8]

In contrast, normal hepatocytes have limited replicative capacity *in vitro* and acquire an abnormal phenotype if they are cultured for extended periods.[9,10] Studies on hepatocytes cultured in a newly devised medium (hepatocyte clonal growth medium[11,12]) revealed a subpopulation of highly replicative PHs, known as small hepatocytes (SHs), in both rats[12] and humans.[13] Their occupancy rate in *h*-liver ranged from 0.01% to 0.09% and was dependent on donor age.[13] The *h*-SHs formed colonies and grew continuously through several subcultures, which led us to name them colony-forming PHs (CFPHs).[13] Replication of the CFPHs was donor age–dependent up to passage 7 ($p = 7$),[13] and the cells did not exhibit a normal hepatocytic phenotype. Instead, they exhibited the traits of hepatocytes or biliary cells depending on the culture conditions. In addition, the CFPHs were not susceptible to infection with hepatitis B virus (HBV) (unpublished data).

In this study, we generated gene expression profiles of CFPHs and transplanted serially subcultured CFPHs into homozygous uPA/SCID mice to examine their growth and differentiation capacity. Our results indicate that the cells were engrafted onto the liver parenchyma and repopulated the tissue, ultimately differentiating into mature hepatocytes. Importantly, the *in vitro*–propagated CFPHs became susceptible to infection with HBV. This study supports our previous suggestion that CFPHs from *h*-postnatal liver are hepatic progenitor-like cells with the potential to assume a normal hepatocytic phenotype.[13]

Materials and Methods

h-*Hepatocytes.* This study was performed with the approval of the Hiroshima Prefectural Institute of Industrial Science and Technology Ethics Board. PHs were isolated as described[13,14] from livers donated by a 12-year-old Asian male (12YM) and a 16-year-old Asian female (16YF) according to the guidelines of the 1975 Declaration of Helsinki. Cryopreserved PHs from a 9-month-old Caucasian male (9MM) and a 10-year-old Caucasian female (10YF) were obtained from In Vitro Technologies (Baltimore, MD) and BD Biosciences (San Jose, CA), respectively.

Culture of CFPHs. Cryopreserved PHs from the 9MM, 12YM, and 16YF were thawed[5] and serially subcultured to obtain *in vitro*–expanded CFPHs.[13] Commercial 9MM PHs and freshly isolated 12YM and 16YF PHs were each subcultured to $p = 3$. The expanded cells were then cryopreserved, thawed upon use, and cultured on collagen-coated plates for 14-20 days as described.[13]

Flow Cytometry. We detached 12YM CFPHs ($p = 4$ or 5) from culture plates by treatment with 0.25% Trypsin-EDTA (Invitrogen, Carlsbad, CA), suspended, incubated on ice for 30 minutes with *m*-monoclonal antibodies against *h*Thy-1 (clone F15-42-1; Chemicon, Temecula, CA), and incubated with antibodies against *m*-immunoglobulin G Alexa-488 (Molecular Probes, Eugene, OR). We used *m*-immunoglobulin G_1 as a negative control. The cells were then analyzed and separated using a fluorescence-activated cell sorter (Becton Dickinson, Franklin Lakes, NJ) as reported.[12]

Transplantation of PHs and CFPHs. We detached 9MM and 12YM CFPHs ($p = 4$) from their culture plates and treated for 1 hour with DMEM containing 10% fetal bovine serum and 3 μg/mL anti–*h*-integrin $\alpha 1$ monoclonal antibodies (clone FB12, Chemicon).[15] This procedure improved engraftment of the CFPHs in uPA/SCID *m*-liver and reduced host mortality.

Transplantation of PHs and CFPHs was performed as described previously.[5] Homozygous uPA/SCID mice were injected with 0.75×10^6 9MM and 12YM PHs or $0.75\text{-}1.0 \times 10^6$ *in vitro*–expanded 9MM and 12YM CFPHs into the inferior splenic pole. When necessary, 10 mM 5-bromo-2′-deoxyuridine (BrdU) (Sigma, St. Louis, MO) and 1.2 mM 5-fluoro-2′-deoxyuridine (Wako, Osaka, Japan) in saline were injected intraperitoneally into the mice at 10 μL/g body weight 1 hour prior to death. The animals were treated according to the guidelines of our local committee on animal experiments.

Table 1. Summary of CFPH and PH Transplantation Experiments in uPA/SCID Mice

Group	Donor Cells	Time of Sacrifice (Weeks After Transplantation)	No. of Transplanted Mice	No. of Mice with Engraftment* [RE (%)]	RI† [Mean ± SD (n)]
A	12YM CFPHs (p = 4)	3	9	3 (33)	0.06-0.19% [0.14 ± 0.07% (n = 3)]
B	9MM CFPHs (p = 4)	3	6	4 (67)	0.03-0.05% [0.04 ± 0.01% (n = 4)]
C	9MM PHs	3	3	3 (100)	5.1-19.4% [6.4 ± 2.9% (n = 3)]
D	12YM CFPHs (p = 4)	9-10	27	14 (52)	0.2-27.0% [6.6 ± 8.3% (n = 14)]
E	9MM PHs	10-11	23‡	23 (100)‡	32.6-82.2% [57.4% (n = 2)]
F	12YM PHs	10	6	4 (67)	31.0-77.0% [62.3 ± 23.8% (n = 4)]
G§	12YM CFPHs (p = 4)	17-20	4	ND	ND

Abbreviation: ND, not determined.

*Number of mice whose livers were engrafted with transplanted PHs or CFPHs. The RE was determined via *h*ALB immunohistochemistry on sections prepared from 5 lobes of a liver.

†Ranges of RI of chimeric mice used in each group.

‡Data from Tateno et al.[5]

§Mice from group G were used for HBV infection studies.

We transplanted 9MM and 12YM CFPHs into 6 and 40 uPA/SCID mice, respectively. The mice were then killed 3, 9, or 10 weeks later, depending on the experimental purpose. In a previous report, we used 9MM and 12YM PHs as donor cells.[5] In this study, we used some of the preserved livers from these mice for histological examinations and as sources of RNA for reverse-transcription polymerase chain reaction (RT-PCR) analysis. The mice used in our transplantation experiments were separated into 7 groups (A-G) as shown in Table 1, which includes the rates of engraftment and replacement indices (RIs) of the chimeric mice.

Blood samples (5 μL) were collected periodically after transplantation from the tail veins of the hosts, and the level of *h*-albumin (ALB) in each was determined using a Human Albumin ELISA Quantitation Kit (Bethyl Laboratories, Montgomery, TX) to monitor the growth of the transplanted CFPHs.

RT-PCR. An RNeasy Tissue Kit (Qiagen, Valencia, CA) was used to isolate total RNA from freeze-thawed 9MM and 10YF PHs, cells of the *h*-hepatoma cell line HepG2, and 12YM and 16YF CFPHs (p = 4). RNA was also isolated with Isogen (Nippon Gene, Tokyo, Japan) from the livers of homozygous uPA/SCID mice and mice chimeric for 12YM PHs or 12YM CFPHs. Each RNA sample was treated with deoxyribonuclease (Takara Bio, Kyoto, Japan) and used as the template for RT-PCR. The RNA (1 μg) was reverse-transcribed with random hexamers using PowerScript Reverse Transcriptase (Clontech, Kyoto, Japan). All reactions were performed with Ex Taq (Takara Bio). Semiquantitative PCR was performed to allow linear amplification of the targets. The following *h*-specific or *m* and *h* cross-reactive genes were subjected to RT-PCR under the conditions shown in Supplementary Table 1: ALB, α1-antitrypsin (AAT), tryptophan-2,3-dioxygenase (TO), glucose-6-phosphatase (G6P), α-fetoprotein (AFP), cytokeratin 19 (CK19), biliary glycoprotein (BGP), Thy-1, CD44, multidrug resistance protein 1 (MDR1), multidrug resistance-associated protein 1 (MRP1), MRP2, and glyceraldehyde-3-phosphate dehydrogenase.

In Situ *Hybridization.* Cryosections (7 μm thick) were fixed with 4% paraformaldehyde, then incubated with 100 ng/mL proteinase K for 10 minutes at 37°C. The sections were then treated at 90°C for 6 minutes and hybridized for 2 hours at 37°C with biotinylated h-DNA probes (Dako, Glostrup, Denmark). The sections were also used to detect whole *h*-genomic DNA using the GenPoint System (Dako) according to the manufacturer's instructions. Finally, they were stained with hematoxylin-eosin.

Immunohistochemistry and Histochemistry. Formalin-fixed livers were embedded in paraffin and sectioned 5 μm thick. The sections were heated in a microwave oven for 5 minutes in Target Retrieval Solution (Dako), then placed at room temperature for 20 minutes. The livers used to generate frozen sections were embedded in OCT compound (Sakura Finechemicals, Tokyo, Japan), frozen in liquid nitrogen, and sectioned 5 μm thick. The cultured cells were fixed in cold ethanol for 10 minutes. The primary antibodies and conditions used for immunohistochemistry are listed in Supplementary Table 2. For bright-field immunohistochemistry, the antibodies were visualized using a Vectastain ABC Kit (Vector Laboratories, Burlingame, CA) using DAB substrates. Fluorescense immunohistochemistry was performed using Alexa 488–conjugated or Alexa 594–conjugated secondary antibodies (Molecular Probes). The nuclei were stained with Hoechst 33258. Glycogens were visualized using a periodic acid-Schiff (PAS) staining kit (Muto Pure Chemicals, Tokyo, Japan). RIs were determined using

*h*ALB-immunostained sections of chimeric *m*-livers as reported previously.[5]

HBV Infection. We obtained *h*-serum containing high-titer HBV DNA (8.1 $\log_{10}$ genome equivalents/mL serum) from an HBV genotype C carrier after obtaining informed consent. The serum was kept at −80°C until use. Four CFPH-chimeric mice were intravenously injected with 100 μL of the HBV-positive serum 9-12 weeks after transplantation.

HBV Marker Analysis. Hepatitis B surface antigen (HBsAg) was measured using an Architect Analyzer (Abbott, Osaka, Japan). Serum DNA was extracted using a SMITEST EX-R&D Nucleic Acid Extraction Kit (Genome Science Laboratories, Fukushima, Japan). Small amounts of HBV DNA (<300 copies/mL) were detected via nested PCR.[8] If HBV DNA was detected during the initial round of PCR, the copy number was determined via real-time PCR as reported.[8]

Results

***Phenotypes of CFPHs* In Vitro.** We seeded 9MM and 12YM PHs on culture dishes and confirmed that the CFPHs from the 2 donors were similar in morphology and replicative capacity. A small number of the CFPHs (0.01%-0.09% of the seeded PHs) began to replicate after 5 days, and the number of replicating cells gradually increased until colonies appeared at 17 days (Fig. 1A); after 21 days, the cells covered the surface of the dish (Fig. 1B). Most of the seeded PHs were not replicative, and they gradually flattened, acquiring a senescent morphology within 20 days of seeding (Fig. 1A). The CFPHs showed an epithelial cell–like morphology with scant cytoplasm (Fig. 1B), and they retained this appearance during subculture (Fig. 1C). The population doubling time (PDT) of the CFPHs gradually increased as the passage number increased. Up to p = 4, the CFPHs from the young donors replicated with a population doubling time of 170-220 hours; subsequently, the population doubling time increased until the cells finally became senescent.[13]

The expression of several marker genes was compared among PHs, HepG2 cells, and CFPHs (Fig. 1D). In our experience, no significant differences exist in the marker gene expression profiles of PHs among different donors, and the same trend applies to subcultured CFPHs.[13] At p = 4, the CFPHs expressed less ALB and AAT messenger RNA compared with the PHs. The PHs expressed TO and G6P, both of which are markers of mature hepatocytes, whereas the CFPHs did not. CK19, a hepatic progenitor/biliary cell marker, was expressed in both the CFPHs and HepG2 cells, but not in the PHs. BGP, a cell–cell adhesion molecule in epithelium, endothelium, and myeloid cells,[16] was expressed in the PHs and HepG2 cells, but only faintly in the CFPHs. The CFPHs, but not the PHs or HepG2 cells, expressed Thy-1, a hematopoietic/hepatic progenitor cell marker. AFP, a hepatic progenitor/carcinoma cell marker, was only detectable in HepG2 cells. CD44, an SH[17] or oval cell marker,[18] was strongly expressed in CFPHs, but only faintly in PHs and HepG2 cells. PHs and CFPHs faintly expressed MDR1. PHs expressed MRP2, but not MRP1. In contrast, CFPHs expressed MRP1, but not MRP2. A change from MRP2 to MRP1 expression during culture has been reported in rat hepatocytes.[19]

Thy-1 and CD44 expression in CFPHs was assessed via immunocytochemistry (Fig. 1E-F). A few CFPHs were positive for Thy-1 (Fig. 1E), whereas the majority was strongly positive for CD44 (Fig. 1F). Fluorescence-activated cell sorting indicated that a minor population of the CFPHs expressed Thy-1 (Fig. 1G-H), with an occupancy rate of 1%-3% (Fig. 1H). The CFPHs expressed CK7, CK8, CK18, and CK19 in the preconfluent state and became CK7- and CK19-negative in condensed regions postconfluence (data not shown), which is in agreement with our previous findings.[13] Other hepatic stem cell markers such as CD34 and c-kit were undetectable in our CFPHs (data not shown).

Repopulation of CFPHs in uPA/SCID Mouse Liver. We transplanted 12YM CFPHs (p = 4) into 27 homozygous uPA/SCID mice. The serum concentration of *h*ALB was monitored posttransplantation as a measure of the RI of CFPHs (Fig. 2A). Approximately half of the hosts had no or only a small increase in the level of *h*ALB throughout the experimental period. The remaining mice showed a continuous increase in the concentration of *h*ALB, which reached >10 μg/mL after 9 to 10 weeks. Animal 27 showed the greatest increase, reaching 0.7 mg/mL after 10 weeks. The RI of each of the 14 mice in which blood *h*ALB concentration was >8 μg/mL after 9 to 10 weeks was determined by dividing the *h*ALB-positive areas by the entire area measured,[5] and the data were plotted against the corresponding blood *h*ALB concentrations (Fig. 2B). RIs between 0.2% and 27.0% were well correlated with blood *h*ALB concentrations in the 9-728 μg/mL range.

Livers of mice engrafted with the CFPHs were subjected to immunohistochemical staining for *h*ALB (Fig. 3A-D,H) and *in situ* hybridization using *h*-genomic DNA probes (Fig. 3I). *h*ALB-positive cells were visible within 3 weeks posttransplantation as single cells or small clusters consisting of up to 25 cells (Fig. 3A-B). Larger clusters containing 20-450 *h*ALB-positive cells appeared after 9 to 10 weeks (Fig. 3C for animal 2 and Fig. 3D for animals 17 and 27). To detect replicating CFPHs, the mice were

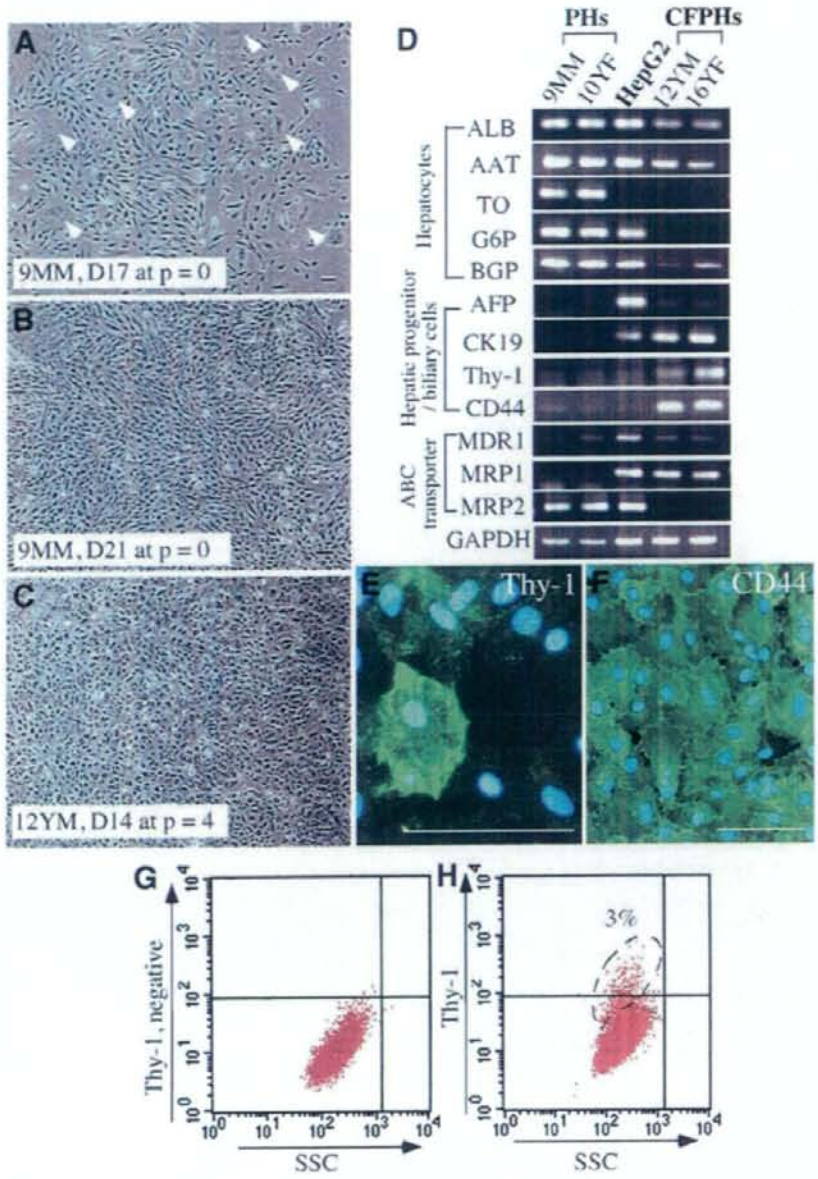

Fig. 1. CFPH growth and gene expression. (A-C) CFPH colony formation. We seeded 9MM PHs at 8×10^3 cells/cm^2 and cocultured with mitomycin C-treated Swiss 3T3 cells in *h*-hepatocyte clonal growth medium. A few CFPHs proliferated and formed colonies. CFPHs were cultured for (A) 17 and (B) 21 days. PHs were nonreplicative and were gradually expelled by replicative CFPHs. Arrowheads indicate the remaining flattened PHs, whose size increased. (C) Cryopreserved 12YM CFPHs (p = 3) were thawed and cultured in *h*-hepatocyte clonal growth medium with Swiss 3T3 cells for 14 days. (D) CFPH messenger RNA expression profiles. RNA was extracted from 9MM and 10YF PHs, HepG2 cells, and 12YM and 16YF CFPHs (p = 4). Semiquantitative RT-PCR was performed for ALB, AAT, TO, G6P, BGP, AFP, CK19, Thy-1, CD44, and the ABC transporters MDR1, MRP1, and MRP2. Glyceraldehyde-3-phosphate dehydrogenase (GAPDH) was used as an internal control. (E,F) Immunohistochemistry of Thy-1 and CD44. 12YM CFPHs (p = 4) were cultured for 14 days and stained for (E) Thy-1 and (F) CD44. The nuclei were stained with Hoechst 33258. Scale bar: 100 μm. (G,H) Flow cytometric analysis of CFPHs for Thy-1. Cells were suspended in Dulbecco's modified Eagle's medium containing 10% fetal bovine serum with (G) *m*-immunoglobulin G_1 as a negative control or (H) anti-*h*Thy-1 antibodies. Living cells were analyzed via fluorescence-activated cell sorting. A small fraction (3% in this case) of the CFPHs was Thy-1$^+$. Three independent analyses were performed with similar results.

given BrdU after 9 weeks. BrdU-positive CFPHs were observed at the edges of the colonies (Fig. 3E-G). Serial liver sections were prepared from CFPH-chimeric mice 9 to 10 weeks after transplantation for *h*ALB immuohistochemistry (Fig. 3H) and for *in situ* hybridization with an *h*-DNA probe (Fig. 3I). The regions identified as containing *h*-hepatocytes by the 2 methods were identical.

Comparison of Repopulation by CFPHs and PHs. PHs and CFPHs (p = 4) were prepared from the livers of 9MM and 12YM donors and transplanted into uPA/SCID mice, and the mice were killed 3 and 10 weeks posttransplantation. The transplanted cells were identified as *h*ALB-positive from histological sections. The number of PH- and CFPH-derived clusters was 125.0 ± 28.2 (n = 3) and 3.3 ± 7.5 (n = 7), respectively, per cross-section of the left lobe of the livers 3 weeks after transplantation, suggesting that the rate of engraftment of the CFPHs was much lower than that of the PHs.

The CFPHs were smaller in size compared with the PHs after 3 weeks (Fig. 4A-B). The cytoplasm of the

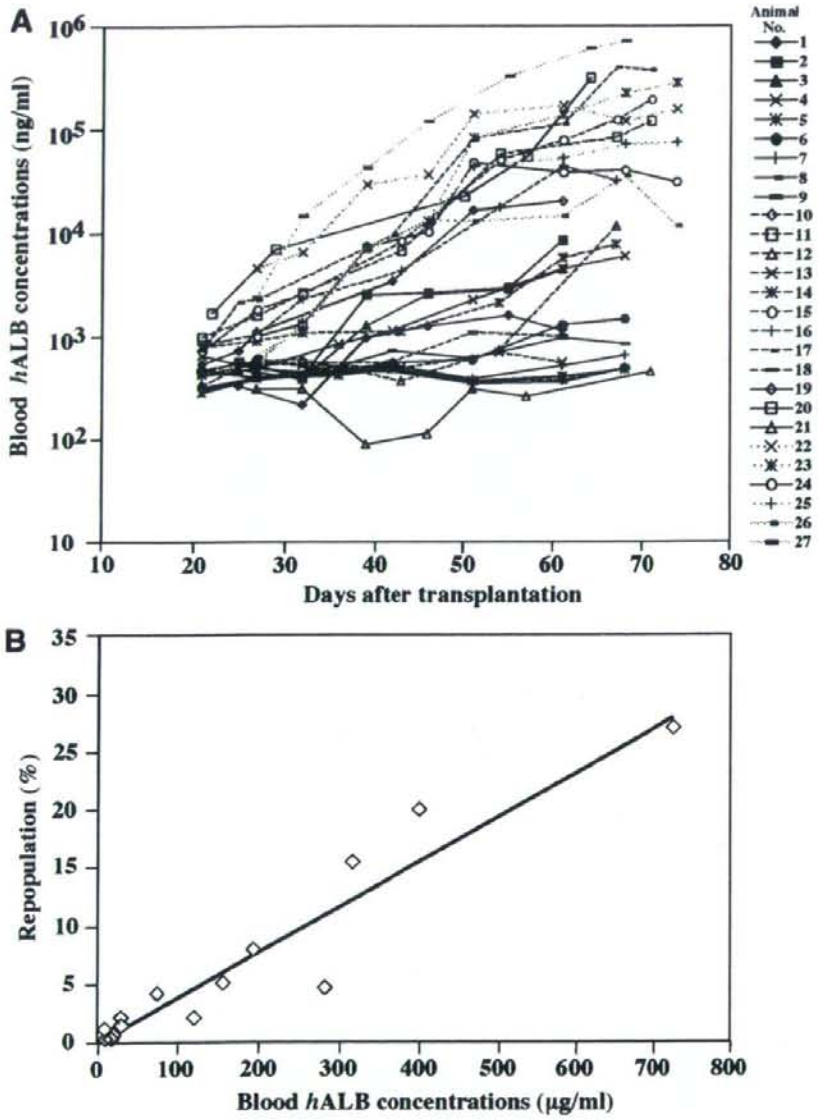

Fig. 2. Transplantation of CFPHs into uPA/SCID mice. The chimeric mice in this experiment are included in group D in Table 1. (A) We transplanted 12YM CFPHs (p = 4) into 27 mice and the serum level of *h*ALB was monitored individually. Ten hosts (animals 1, 5, 6, 7, 8, 9, 10, 13, 18, and 21) did not show significantly elevated *h*ALB levels during the experimental period. Four hosts (2, 3, 4, and 14) showed slight elevation. The *h*ALB concentration of 13 mice (11, 12, 15, 16, 17, 19, 20, 22, 23, 24, 25, 26, and 27) reached >10 μg/mL at 9 to 10 weeks after transplantation. (B) Correlation between the blood *h*ALB level and RI. Fourteen CFPH-chimeric mice (animals 2, 11, 12, 15, 16, 17, 19, 20, 22, 23, 24, 25, 26, and 27) were selected from the mice shown in panel A for RI determination. Their liver sections were immunostained for *h*ALB. RIs were determined for each animal and plotted against the *h*ALB concentration. The correlation coefficient (r^2) between the 2 parameters was 0.91.

former was less abundant and more strongly stained for *h*ALB than that of the latter. We observed *h*CD44 in the plasma membrane of the CFPH-derived cells (Fig. 4E), but not in that of the PH-derived cells (data not shown). At 10 weeks posttransplantation, the CFPHs had increased in size to match those of the PHs, whose sizes were unchanged (Fig. 4C-D), and *h*CD44 expression disappeared from the CFPH-derived cells (Fig. 4F). The diameter of each CFPH and PH was quantified as follows: 18.3 ± 5.1 μm (mean ± SD, n = 65) versus 25.8 ± 6.4 μm (n = 124) at 3 weeks and 27.0 ± 5.5 μm (n = 185) versus 25.8 ± 4.8 μm (n = 187) at 10 weeks. We found no significant differences in this parameter between the 12YM and 9MM samples. Thus, it appears that the CFPHs replicated without changing their original small size until 3 weeks posttransplantation, when they became larger.

Liver sections from the chimeric mice were stained with hematoxylin-eosin to compare the morphological features of PHs and CFPHs at 10 weeks. The repopulated CFPHs (Fig. 4G) showed no significant difference in morphology compared with the repopulated PHs (Fig. 4H). As reported previously,[5,6] the PHs in the chimeric livers were enlarged and had less eosinophilic cytoplasm

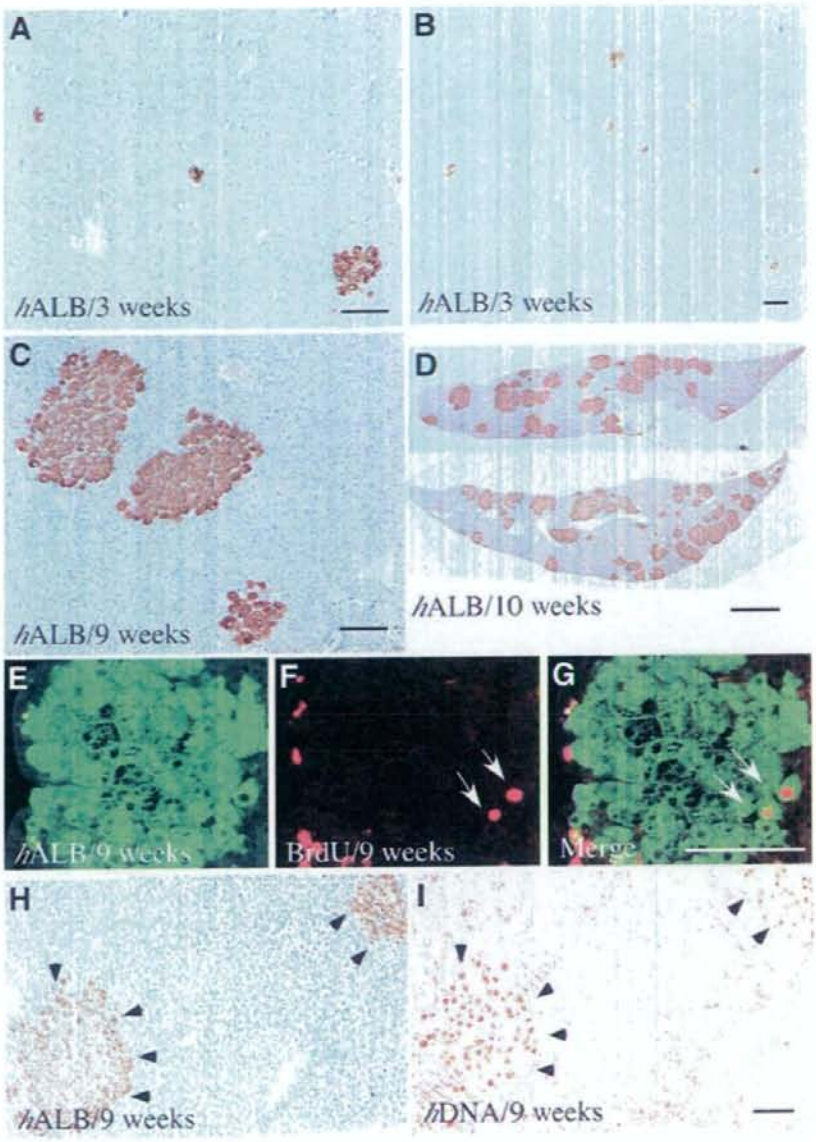

Fig. 3. Engraftment and repopulation of CFPHs in chimeric mouse liver. The chimeric mice in this experiment are included in groups A and D in Table 1. We performed *h*ALB immunohistochemistry using liver sections from CFPH-chimeric mice (A,B) 3, (C) 9, and (D) 10 weeks after transplantation. (A,B) Small clusters composed of 1-25 cells were scattered throughout the liver at 3 weeks in 3 of 9 mice. (C,D) The clusters became larger at 9 to 10 weeks. The liver sections in panel C were prepared from animal 2 in Fig. 2A (RI = 1.1%). The liver sections in panel D were prepared from animals 17 (RI = 20.0%; upper section) and 27 (RI = 27.0%; lower section). Three mice were randomly selected for the BrdU incorporation experiments (animals 2, 19, and 20 in Fig. 2A). They were given BrdU 1 hour before death at 9 weeks posttransplantation. Serial liver sections were subjected to (E) *h*ALB- and (F) BrdU immunohistochemical staining. The image in panel G is panel E and panel F merged. Similar results were obtained from these experiments, and the result from animal 19 (RI = 0.6%) is shown in panels E-G. Serial liver sections were prepared from CFPH-chimeric mice (animals 2, 15, and 17 in Fig. 2A) 9 to 10 weeks after transplantation for *h*ALB immunohistochemistry (H) and for *in situ* hybridization with an *h*-genomic probe (I). Similar results were obtained from the 3 mice. The results shown in panels H and I were obtained from animal 2 (positive cells are indicated by arrowheads). Scale bars in panels A-C, G, and I: 100 μm. Scale bar in panel D: 1 cm.

than the PHs in *h*-livers. The livers of the mice that had low *h*ALB levels at 10 weeks posttransplantation were mostly occupied by red nodules, which have been reported to be formed by the transgene-deleted hepatocytes of the host.[20]

Gene and Protein Expression Profiles of CFPHs in Chimeric Mice Compared with Those of PHs. Three 12YM CFPH-chimeric mice (11, 15, and 17) were randomly selected from the mice in Fig. 2A and killed 10 weeks after transplantation. RNA was extracted from each liver to generate gene expression profiles via RT-PCR. RT-PCR was also performed on 2 12YM PH-chimeric mice that were included in a previous study.[5] The CFPH livers expressed *h*ALB, *h*AAT, *h*TO, *h*G6P, and *h*MRP2, but not *h*CK19, *h*Thy-1, or *h*MRP1, just as in the PH-livers (Fig. 5). Previously, we showed that the PHs in chimeric mice expressed various *h*-cytochrome P450 (*h*CYP) subtypes in a manner similar to the donor liver.[5] In this study, we found that the expression of *h*CYPs 1A2, 2C8, 2C9, 2D6, and 2E1, but not 3A4, in the CFPH-chimeric mice was similar to that in the PH-chimeric mice (data not shown). Expression of *h*CYP3A4 was very

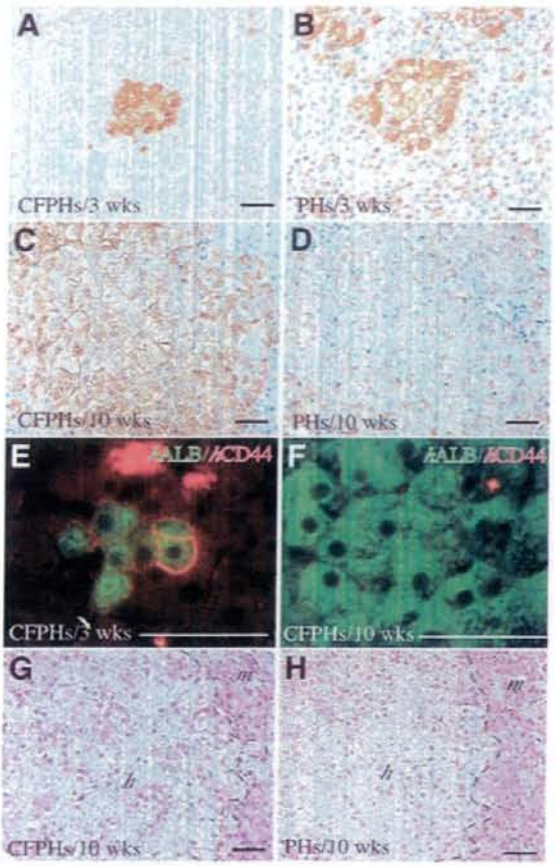

Fig. 4. Immunohistochemical staining for CFPHs and PHs in chimeric mice. Immunohistological analysis with antibodies against (A-D) *h*ALB and (E-F) *h*CD44. We produced 3 12YM CFPH-chimeric mice and 4 9MM CFPH-chimeric mice [(A) and (E), included in groups A and B in Table 1] and 3 9MM PH-chimeric mice [(B), group C], which were killed at 3 weeks posttransplantation. At 10 weeks posttransplantation, 3 12YM CFPH-chimeric mice that were randomly selected from the mice shown in Fig. 2A (15, 16, and 17) were killed [(C) and (F), group D], as were 9MM and 12YM PH-chimeric mice, 2 mice each [(D), groups E and F]. (A-D) Representative images of liver sections prepared from the animals and stained with anti-*h*ALB antibodies. The diameters of the *h*ALB-positive cells were measured in 10-15 randomly selected fields. (E,F) Double-fluorescence immunostaining. Green and red stains depict *h*ALB and *h*CD44, respectively. (G,H) Hematoxylin-eosin staining. (G) Eight CFPH mice were randomly selected from the mice shown in Fig. 2A and killed at 10 weeks posttransplantation. Their liver tissues were then subjected to hematoxylin-eosin staining. (H) Three 12YM PH-chimeric mice were killed at 10 weeks posttransplantation for hematoxylin-eosin staining as above. Similar results were obtained for the 8 CFPH-chimeric mice and 3 PH-chimeric mice. (E-F) Sections from (E) a CFPH-chimeric mouse (RI = 20.0%) and (F) a PH-chimeric mouse (RI = 57%). *h*, *h*-hepatocyte region; *m*, *m*-hepatocyte region. Dashed lines show the boundary between the 2 regions. Scale bars: 50 μm.

low (less than one-fifth) in CFPHs compared with that in PHs.

Protein expression was investigated immunohistochemically for the CFPH-chimeric livers at 3, 9, and 10 weeks posttransplantation. All of the examined CFPHs were Thy-1–negative, CK7-negative, CK19-negative, and AFP-negative (data not shown). The *h*ALB-positive cells were coincident with the *h*CK18-positive cells at both 3 (data not shown) and 9 weeks posttransplantation (Fig. 6A-C). MRP2-positive signals were present on the bile canalicular membranes of the transplanted CFPHs at 10 weeks (Fig. 6D-F). CYP3A4-expressing CFPHs were localized in the pericentral zone (Fig. 6G-I) as reported previously,[21] but their distributions were unique. Although some of the CFPHs were positive for CYP3A4, approximately 70% of them were negative. In contrast, all of the CFPHs in the pericentral zone strongly expressed CYP1A2 (Fig. 6J-L), which is known to be expressed in postnatal liver.[22] The CFPHs in the chimeric mice were strongly PAS-positive (Fig. 6N), whereas the *in vitro* CFPHs were faintly PAS-positive (data not shown). From

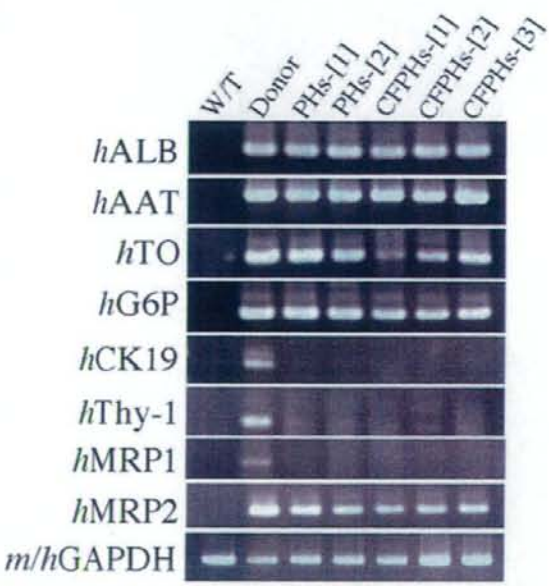

Fig. 5. Gene expression profiles of CFPHs in chimeric mice. Two uPA/SCID mice were transplanted with 12YM PHs ([1] and [2]); 3 uPA/SCID mice were transplanted with 12YM CFPHs ([1], [2], and [3]). The chimeric mice in this experiment are included in groups D and F in Table 1. After 10 weeks, the livers were removed for RT-PCR analysis. At the time of death, the PH-[1]-, PH-[2]-, CFPH-[1]-, CFPH-[2]-, and CFPH-[3]-chimeric mice had RIs of 41.0%, 57.0%, 2.1%, 7.9%, and 20.0%, respectively. The analysis was repeated using liver tissues from donor and uPA/SCID mice without transplantation (W/T). Glyceraldehyde-3-phosphate dehydrogenase (GAPDH) amplification was used as an internal control.

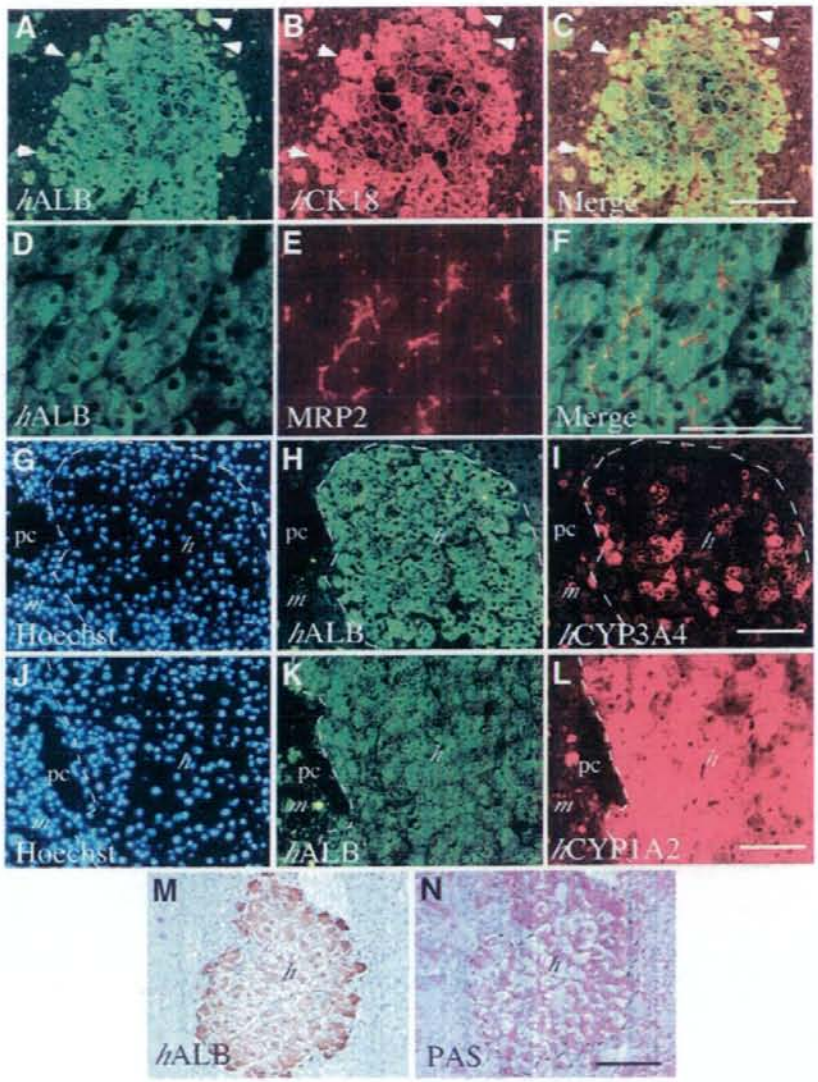

Fig. 6. Protein expression profiles of the CFPHs in chimeric livers. Mice were transplanted with 12YM CFPHs, and their livers were removed 9 to 10 weeks after transplantation for immunohistochemical analysis of (A,D,H,K) *h*ALB, (B) *h*CK18, (E) MRP2, (I) CYP3A4, and (L) CYP1A2. The chimeric mice in this experiment are included in group D in Table 1. Representative images are shown. (A-F) Double-fluorescence immunostaining. (A,D) *h*ALB is stained green. (B) *h*CK18 and (E) *h*MRP2 are stained red. Panels A and B were merged to create panel C; panels D and E were merged to create panel F. The arrowheads in panels A-C show macrophages engulfing such wastes as lipids. Serial sections of liver tissues subjected to 2 series of immunohistochemical examinations, one for (G-I) *h*CYP3A4 and the other for (J-L) *h*CYP1A2. The sections were stained with (G,J) Hoechst 33258, and for (H,K) *h*ALB, (I) *h*CYP3A4, and (L) *h*CYP1A2. Serial sections of liver tissues at 9 weeks posttransplantation were subjected to *h*ALB-immunostaining (M) and PAS staining (N). The positive cells appear brown in (M) and red in (N). *h*, *h*-hepatocyte region; *m*, *m*-hepatocyte region; pc, pericentral zone. Dashed lines show the boundary between the *h*-hepatocyte and *m*-hepatocyte regions. Scale bars: 100 μm.

these results, we conclude that the transplanted CFPHs differentiated into functionally mature hepatocytes. No *h*-cell tumors were formed during any of our experiments in the uPA/SCID mice.

Infection of CFPH-Chimeric Mice with HBV. To further examine whether CFPHs had exhibited normal differentiated phenotypes in chimeric mice, we tested their susceptibility to HBV infection. Four CFPH-chimeric mice with various serum *h*ALB levels (0.2, 1.6, 7.3, and 222.0 μg/mL) were inoculated with 100 μL of HBV-positive *h*-serum at 9-12 weeks posttransplantation. The animals were then tested every 2 weeks for HBV viremia and serum *h*ALB levels (Fig. 7A). The amount of HBV DNA in the animals increased between 2 and 8 weeks after inoculation, and all 4 mice developed measurable viremia within 8 weeks. However, a correlation was observed between the HBV DNA and/or HBsAg level and the *h*ALB level: the former appeared to be high when the latter was high (Fig. 7A). HBsAg was detectable in the serum of the chimeric mice when they showed elevated virus titers: the HBsAg levels of chimeric mice with HBV DNA levels of 2×10^3, 5.2×10^5, 5.9×10^7, and 7.7×10^8 copies/mL 8 weeks after inoculation were <0.05, <0.05, 3.2, and 124.0 IU/mL, respectively. HBV was infectious to CFPH-chimeric mice with very low levels of *h*ALB ($<10^4$ ng/mL), and all mice showed quantitatively

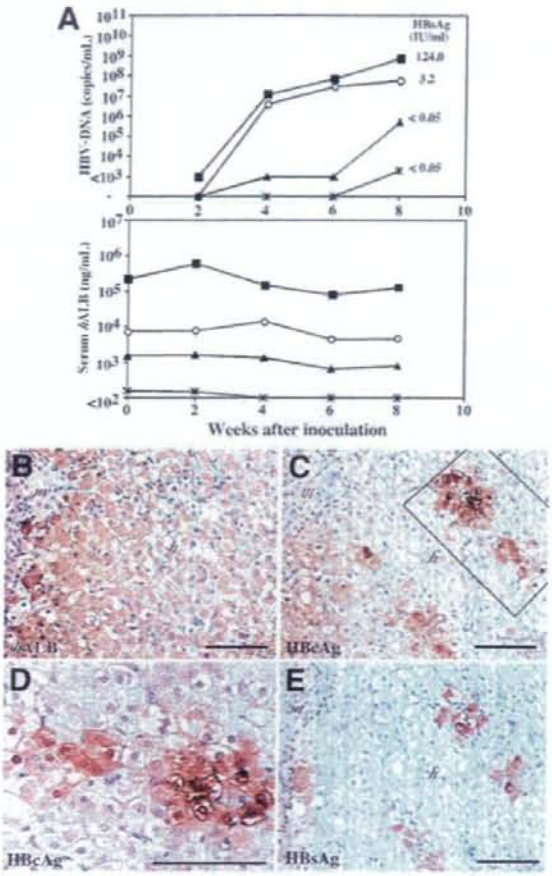

Fig. 7. Susceptibility of chimeric mice to infection with HBV. The chimeric mice in this experiment are included in group G in Table 1. uPA/SCID mice were transplanted with 12YM CFPH (p = 4). (A) The serum *h*ALB concentration of each mouse was determined 9-12 weeks posttransplantation just before the mouse was intravenously injected with 100 μL of HBV-positive *h*-serum (0.2 μg/mL at 12 weeks, 1.6 μg/mL at 10 weeks, 7.3 μg/mL at 11 weeks, and 222.0 μg/mL at 9 weeks). The animals were examined every 2 weeks for HBV viremia and serum *h*ALB level. The upper and lower graphs show the HBV DNA levels (copies/mL) and serum *h*ALB concentrations (ng/mL), respectively. The amount of HBV DNA ($<10^3$ copies/mL) was semiquantitatively measured via nested PCR. The values in the upper graph represent the HBsAg levels at 8 weeks. (B-E) Immunohistochemical analysis of chimeric livers infected with HBV. Serial sections of liver tissues at 8 weeks after inoculation were stained for (B) *h*ALB, (C,D) hepatitis B core antigen, and (E) HBsAg. The region enclosed by a square in panel C is magnified in panel D. Scale bars: 100 μm.

measurable viremia ($>10^3$ copies/mL) up to 8 weeks after inoculation. In contrast, most PH-chimeric mice with $<10^4$ ng/mL *h*ALB did not show quantitatively measurable levels of viremia up to 12 weeks after inoculation (data not shown) as reported previously.[8] In this study, we confirmed that CFPHs were not susceptible to infection with HBV prior to transplantation. The presence of hepatitis B core antigen and HBsAg in the CFPHs from HBV-infected chimeric livers was examined immunohistochemically (Fig. 7C,E). CFPHs were positive for both antigens that were sporadically distributed in the same regions among the CFPH colonies. Hepatitis B core antigen–positive cells accounted for 18.7 ± 8.3% of the total number of CFPHs (n = 3; total cell count = 1,215) (Fig. 7C), and both the nucleus and cytoplasm of the cells showed signals (Fig. 7D).

Discussion

This study supports our previous conclusion that CFPHs are *h*-hepatic progenitor-like cells.[13] Cultured CFPHs expressed such hepatic progenitor cell markers as CK19, Thy-1, and CD44, but not mature hepatocyte markers such as TO and G6P. We also found that *in vitro*–expanded CFPHs in uPA/SCID mice were able to repopulate the parenchyma, in which they differentiated into mature hepatocytes. FISH (fluorescence *in situ* hybridization) using mouse X chromosome probes showed that the engrafted and propagated CFPHs did not fuse to the mouse cells (data not shown). Thus, replicative CFPHs isolated from postnatal liver are normal, functional hepatocyte progenitor-like cells.

The existence of stem/progenitor cells in the adult liver is controversial.[23-25] In the present study, we showed that the CFPHs expressed CK19, Thy-1, and CD44, but not AFP, in serial culture. Thy-1 antigens are expressed in *h*-hepatic progenitor cells in fetal liver[26] and in rat oval cells,[27] but not in normal adult hepatocytes. We showed that Thy-1-expressing cells were present among the CFPHs at an occupancy of 1%-3%. SHs show greater growth potential than PHs in rats.[12] Other studies have reported that CD44 is a specific marker for rat SHs *in vitro* and *in vivo*, and that its expression level decreases with SH maturation *in vitro*.[17] Moreover, a recent study demonstrated that CD44 was strongly expressed by oval cells in a 2-acetylaminofluorene/partial hepatectomy, a D-galactosamine, and a retrorsine/partial hepatectomy rat model, but not by small hepatocyte-like progenitor cells (SHPCs)[18] that appeared in a retrorsine/partial hepatectomy model.[28] We detected CD44 expression in CFPHs at the plasma membrane. These results suggest that Thy-1 and CD44 may be common markers for both rat and *h*-hepatic progenitor cells.

Mouse embryonic liver stem cell lines differentiate into both hepatocytes and bile ducts in uPA/SCID mice.[29] Like PHs, our CFPHs differentiated into mature hepatocytes, but not into biliary epithelial cells, in uPA/SCID mice. CFPHs are considered to be hepatic progenitor-like cells, like rat SHs[12,30-33] and SHPCs.[28,34] SHPCs are

closely related to SHs; they are small and similar in size,[28,30] and both express CYP3A1 and 2E1 at a low level.[28,32] At 3 weeks posttransplantation, the CFPHs were small in size, had a large nucleus-to-cytoplasm ratio, and expressed *h*CD44, but not *h*CK19. At 10 weeks, the cells became bigger, assumed a morphology similar to that of PH-derived cells, and lost their expression of *h*CD44. The expression of *h*CYP3A4 was quite low (0.15-fold) among CFPHs compared with that of PHs (data not shown). In addition, the distribution of *h*CYP3A4-expressing CFPHs in the pericentral zone was unique: more than two-thirds of CFPHs did not express CYP3A4. In the case of the *h*-PH–chimeric mice, all PHs in the pericentral zone expressed CYP3A4 (data not shown).

Presently, we lack experimental data to explain the expression of *h*CYP3A4 in CFPH-chimeric liver, but CFPHs may require some specific environmental factor(s) for differentiation, which might be absent from mouse liver. Alternatively, some factors that specifically inhibit the differentiation of CFPHs might be present there. CK7-positive *h*-hepatic progenitor cells are present in the livers of uPA/SCID mice transplanted with *h*-postnatal liver-derived PHs,[6] and these small cells are strongly immunoreactive to pan-cytokeratin with scant cytoplasm. The CFPHs were morphologically similar to these cells at 3 weeks posttransplantation, although we were unable to detect CK7-positive cells in either the PH- or CFPH-transplanted chimeric livers. However, CFPHs were *h*CK7-, *h*CK19-, and *h*CD44-positive, at least until 1 day posttransplantation (data not shown).

We reported previously that uPA/SCID livers were nearly completely replaced with young donor PHs at 10 weeks posttransplantation.[5] In contrast, the RIs of our CFPH-chimeric mice were <30% at 9 to 10 weeks. CFPHs were rare in the host liver at 3 weeks posttransplantation, whereas several PHs were observed. The lower RIs of the CFPHs might be attributable to their lower engraftment efficiency.

In conclusion, *h*-hepatocytes in immunodeficient, and liver-injured mice are useful for the study of viral hepatitis. Repopulated *h*-hepatocytes are susceptible to infection with HBV[6-8] and HCV.[4,6] Additionally, *h*-hepatocyte–chimeric mice are usually produced by transplanting fresh[6,7] or cryopreserved hepatocytes,[4,5] but sources of *h*-hepatocytes are limited. Several studies have reported on liver repopulation by *in vitro*–propagated cells from adult and fetal livers, such as immortalized mouse hepatic stem cells,[29] rat SHPCs,[34] immortalized *h*-hepatocytes transfected with full-length HBV,[35] and fetal *h*-epithelial/hepatic progenitor cells.[36,37] However, the RIs in these studies were extremely low (less than a few percent). In the present study, we were able to produce CFPH-chimeric mice with RIs as high as 27%. Thus, CFPHs could be an alternative to *h*-hepatocytes as a source of hepatocytes for transplantation. Moreover, the CFPH-chimeric mice were susceptible to infection with HBV, even though their serum *h*ALB levels were extremely low (10^2-10^3 ng/mL). CFPH-chimeric mice will be useful for studying *h*-HBV and for characterizing *h*-hepatic progenitor cells.

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